**Exploring the Antibacterial Potential of Essential Oils Extracted from Three Medicinal Plants Against Some Foodborne Bacteria**

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**ABSTRACT**

This study aims to evaluate the antibacterial activity of essential oils extracted from the green leaves of three medicinal plants, namely Cupressus macrocarpa, Schinus terebinthifolius, and Eucalyptus citriodora, against selected foodborne bacteria. Gas chromatography/mass spectrometry (GC/MS) analysis was employed to identify the chemical composition of the extracted essential oils. The two main chemical components of C. macrocarpa essential oils (EO) were terpinene-4-ol (32.37%) and citronellol (29.29%). The primary components of S. terebinthifolius EO were α-phellandrene (44.35%) and o-cymene (10.42%). Meanwhile, α-phellandrene (13.5%) and sabine (24.24%) constitute the majority of E. citriodora EO. The antibacterial activity of the essential oils was assessed against Gram-positive bacteria including Bacillus cereus, Staphylococcus saprophyticus, Lysinibacillus fusiformis, and Kocuria rhizophila, as well as the Gram-negative bacterium Serratia liquefaciens. Standard antibiotics like Penicillin, Amoxicillin, and Ampicillin were used for comparison. The results revealed varying degrees of antibacterial activity against each pathogenic isolate used. C. macrocarpa essential oil exhibited the strongest antibacterial action, with a 55.7 mm inhibition zone diameter against Bacillus cereus, which displayed resistance to the tested standard antibiotics. Meanwhile, essential oils of the chosen plants also inhibited the growth of S. liquefaciens despite its tolerance to the tested antibiotics. The minimum inhibitory concentrations (MIC) of C. macrocarpa, S. terebinthifolius, and E. citriodora essential oils ranged from 0.06 to 1.5 mg/ml, 0.68 to 2.0 mg/ml, and 0.2 to 1.77 mg/ml, respectively. These findings highlight the potential of the tested essential oils as antibacterial agents for preserving food materials in a safe, sustainable, cost-effective, and eco-friendly manner.

**Keywords:** Antibacterial activity, foodborne bacteria, Food preservation; Essential oils, Medicinal plants.

**INTRODUCTION**

Essential oils (EO) derived from medicinal plants are valuable secondary metabolites widely utilized in the food industry for their fragrance and antimicrobial properties (Serag et al., 2022). Owing to their natural origin and inherent protective properties in plants, essential oils have garnered significant interest as potential antimicrobial agents (Gunasena et al., 2022). These oils are composed of aromatic volatile compounds obtained from various parts of plants (Wang et al., 2020). Microbial contamination poses a hidden risk to food safety, making the exploration of effective antimicrobial strategies crucial.

The utilization of synthetic antimicrobial agents has proven to be an effective approach in inhibiting microbial growth, reducing the risk of food poisoning, and extending the shelf life of food products (Shatalov et al., 2017). The food industry faces significant challenges due to the contamination of food with pathogenic microorganisms, such as bacteria and fungi, which produce toxins that degrade food quality during pre- and postharvest processing (Maurya et al., 2021). To address this issue, chemical preservatives have been employed, including formaldehyde, sodium benzoate, pyrrolidines, sulfites, sodium nitrite, sulfur dioxide, benzoates, imidazoles, thiocyanates, and sorbates, demonstrating their efficacy in reducing microbial contamination in food (Gutiérrez-del-Río et al., 2018).

In light of their potential human health risks, including carcinogenesis, teratogenesis, environmental toxicology, and long-term degradation (Falleh et al., 2020), the use of chemical preservatives mentioned earlier raises concerns about negative health impacts. In addition, these synthetic preservatives can also accumulate in the environment, leading to pollution and potential harm to ecosystems (Falleh et al., 2020). Furthermore, the emergence of antibiotic-resistant foodborne pathogens poses a significant threat to public health and food security. The misuse or overuse of antibiotics in both human medicine and agriculture has contributed to the development of antibiotic resistance among bacterial pathogens (Bhatia et al., 2021). This has led to the need for alternative strategies to combat these pathogens and ensure the safety of our food supply. Therefore, the development of natural preservatives has emerged as an alternative approach (Marrone et al., 2021). To combat this issue, numerous studies have highlighted the potent antibacterial properties of plant essential oils (Isepí et al., 2019). These natural alternatives show promise in overcoming the challenges associated with synthetic antimicrobial agents and chemical preservatives, contributing to the safeguarding of food safety and public health.

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The utilization of natural plant extracts in the field of food preservation has gained significant attention. *Cupressus macrocarpa*, a medicinal plant belonging to the Cupressaceae family, possesses various pharmacological properties such as hepatoprotective, anti-inflammatory, and antiviral activities (Al-Sayed et al., 2018; Negm et al., 2020). The essential oil derived from *C. macrocarpa* has been found to exhibit potent antibacterial and antifungal properties, effectively targeting a wide range of bacteria and fungi (Attallah et al., 2021). Another plant of interest is *Schinus terebinthifolius* Raddi, a perennial woody plant from the Anacardiaceae family (Belhoussaine et al., 2022). This plant has been associated with various pharmacological activities, including anti-inflammatory effects (Silva et al., 2017), antimicrobial properties (da Silva et al., 2018), and antioxidant activity (Rocha et al., 2018). *Eucalyptus citriodora* Hook, a member of the Myrtaceae family (Valenzuela et al., 2021), has also demonstrated diverse activities, including antibacterial, antifungal, and pharmaceutical properties (Gu et al., 2019). Such characteristics make *E. citriodora* a promising candidate for natural preservation methods in the food industry. The exploration of these medicinal plants and their essential oils provides potential alternatives to synthetic antimicrobial agents and chemical preservatives. By harnessing the antibacterial and antifungal properties of *Cupressus macrocarpa*, *Schinus terebinthifolius*, and *Eucalyptus citriodora*, researchers aim to develop natural preservatives that can effectively inhibit microbial growth, reduce food contamination, and preserve food quality.

Regarding foodborne pathogens, *S. saprophyticus* is a common contaminant found in meat, poultry-contaminant and fermented foods, as well as in the intestinal and rectal flora of animals like pigs and cattle (Becker et al., 2014). This bacterium is notorious for causing urinary tract infections, primarily by colonizing the gastrointestinal tract and subsequently being inadvertently transferred to the urethra through contaminated feces (Sommers et al., 2017). Another significant foodborne pathogen is *Bacillus cereus*, which has been associated with outbreaks worldwide (Elaffify et al., 2023). Meanwhile, *Lysinibacillus fusiformis*, a rod-shaped Gram-positive bacterium, poses a threat to human health, causing tropical ulcers, severe sepsis, and respiratory illnesses (Sulaiman et al., 2018). Additionally, *Kocuria* organisms, belonging to the Gram-positive coccoid bacteria group in the Micrococaceae family, can cause various infections, especially in individuals with compromised immune systems and underlying disorders (Savini et al., 2010). Notably, the recent reclassification of the ATCC 9341 strain as *Kocuria rhizophila* from *Micrococcus luteus* adds to our understanding (Tang et al., 2003). Finally, *Serratia*, a Gram-negative member of the Enterobacteriaceae family, is recognized for its association with food spoilage. *S. liquefaciens*, a species within this genus, acts as an opportunistic pathogen in humans and is highly pathogenic to humans, insects, and fish (Mahlen, 2011).

The infections caused by *Serratia* species can be severe, even leading to fatality (Samonis et al., 2011; Parte et al., 2020). Given the risks posed by these foodborne pathogens, the development of natural preservatives becomes crucial. Therefore, this study aims to investigate the potential activity of essential oils of *Cupressus macrocarpa*, *Schinus terebinthifolius*, and *Eucalyptus citriodora* as antibacterial against some foodborne bacteria.

**MATERIALS AND METHODS**

**Plant material collection**

Fresh green leaves of *Cupressus macrocarpa*, *Schinus terebinthifolius*, and *Eucalyptus citriodora* were collected from the Botanical Garden of the Faculty of Education, Damietta University, during the Spring-Summer season of 2021. The collected plant samples underwent authentication by Dr. Mamdouh S. Serag, a Professor of Plant Ecology in the Botany and Microbiology Department of the Faculty of Science, Damietta University. To ensure proper documentation and future reference, plant specimens were carefully prepared and deposited in the herbarium of the Botany and Microbiology Department, Faculty of Science, Damietta University. This repository serves as a valuable resource for the verification and further study of the collected plant samples.

**Extraction of essential oils**

Fresh leaves of the selected plants (500 g) were washed with distilled water, segmented into small parts, then added distilled water and heated to boiling using a Cleveger-type apparatus and hydro-distillation for 5 hours. After boiling, the essential oil evaporated, condensed as the upper phase, and separated from the lower one. The extracted oil was dried over anhydrous sodium sulfate and kept in a closed, dark glass vial at 4 °C in the refrigerator until analysis (Boukhris et al., 2012).

**Gas chromatography-Mass spectrometry analysis (GC/MS)**

At the Central Laboratories Network, National Research Centre, Cairo, Egypt, a gas chromatograph (7890B) and mass spectrometer detector (5977A) were part of the GC-MS system (Agilent Technologies). An HP-1MS column (60 m x 0.25 mm internal diameter and 0.25 μm film thickness) was installed in the GC. Helium was used as the carrier gas for the analyses, with a flow rate of 1 ml/min, a split ratio of 30:1, an injection volume of 0.5 μl, and a temperature program of 40 °C for 1 min, followed by a rise of 5 °C/min to 200 °C. The injector and detector were kept at respective temperatures of 250 and 280 °C. By employing an m/z range of 50-550 and a solvent delay of five minutes, mass spectra were produced by electron ionization (EI) at 70 eV. The spectrum fragmentation pattern was compared to those stored in the Wiley and NIST Mass Spectral Library data to identify various constituents.

**Media used for isolation**

Different media were utilized for the isolation of foodborne bacteria, including nutrient agar and Nutrient broth. Nutrient agar, comprising 5.0g peptone, 3.0g beef extract, 8.0g sodium chloride, and 12.0g agar per liter, was purchased from Neogen (Heywood, UK).
for solid medium preparation (Atlas, 2010). Nutrient broth, containing 5.0 g peptone, 3.0 g beef extract, 5.0 g sodium chloride, 5.0 g glucose, and distilled water to achieve a final volume of 1000 ml, was also obtained (Atlas, 2010). Additionally, Mueller Hinton Agar medium, purchased from Britania SA Labs (Buenos Aires, Argentina), was also used, consisting of (g/L): 3.0 beef extract, 17.5 acid casein hydrolysate, 1.5 starches, and 15.0 agar. These media were selected based on their established formulations and suitability for the Isolation and cultivation of foodborne bacteria (Atlas, 2010).

Isolation of bacterial strains

All the tested bacteria were isolated from spoiled food samples, specifically beef burgers, beef lunch- eons, and sausage, in the Microbiology laboratory at the Faculty of Science, Damietta University. The samples were randomly purchased from various markets in New Damietta, Egypt, and were immediately transferred to sterile polyethylene bags to maintain aseptic conditions. These samples were then stored at room temperature to create favorable conditions for spoilage and microbial growth.

Morphological, biochemical, and molecular techniques for Identification of Isolated Bacterial Strains

The isolated bacterial strains were subjected to a comprehensive identification process involving morphological, biochemical, and molecular techniques. Morphological characteristics, such as colony morphology and cell shape, were observed under a microscope. Additionally, Gram staining (McClelland, 2001) and spore staining (Gerhardt, 1994) were performed for further aid in the identification process. The strains were assessed based on their shape, pigment production, elevation, edge, color, and opacity.

Biochemical tests were conducted to gather additional information about the bacterial strains. Carbohydrate fermentation tests and enzyme activity assays were employed to determine the metabolic properties of the isolates. These tests provided valuable insights into the metabolic capabilities of the strains, assisting in the characterization and identification process. In order to achieve higher accuracy and specificity in identification, molecular technique utilizing 16S rRNA was employed. Polymerase chain reaction (PCR) was utilized to amplify the 16S rRNA gene, and subsequent DNA sequencing was performed. This molecular approach allowed for more precise identification of the bacterial isolates. Overall, a combination of morphological, biochemical, and molecular methods was employed to comprehensively identify the isolated bacterial strains.

Biochemical characteristics

Bacterial isolates were identified using Bergey’s manual of determinative bacteriology (Breed et al., 1957; Grimont and Grimont, 2006; and Langlois et al., 1990). Biochemical tests including catalase (Cappuccino and Sherman, 2001), starch hydrolysis, Voges Proskauer, urea hydrolysis, citrate utilization, Hydrogen sulfide (H₂S) production (Atlas, 2010), Sugar fermentation test (MacFaddin, 2000), indole production (Ojokoh and Eromosele, 2015), tube coagulase test (katz et al., 2010), oxidase test (MacFaddin, 2000), and esculin hydrolysis (Atlas, 2010).

Molecular identification of the isolated bacterial strains

DNA extraction

According to the manufacturer’s instructions, genomic DNA was extracted and purified from bacterial samples using ABT DNA mini extraction kit (Applied Biotechnology Co. Ltd, Egypt).

PCR amplification of 16S rRNA

PCR was carried out using the universal primer pair 27F (5’-AGA GTT TGA TCC TGG CTC AG-3’) and 1492R (5’-GTT TAC CTT GTG ACC ACT T-3’). PCR reactions were performed in a volume of 50 µl (2x Red Master mix, 20 picomoles of each primer, and 100 ng of genomic DNA) using a thermal cycler (MJ Research, USA). PCR cycling conditions were as follows: one cycle of initial denaturation stage at 95 ºC for 3 min, followed by 35 amplification cycles [Denaturation at 95 ºC for 30 s, annealing at 50 ºC for 30 s, extension at 72 ºC for 90 s], then a final extension step at 72 ºC for 5 min. PCR-DNA products (5µl) were visualized on ethidium bromide-stained 1% agarose gel (w/v) in Tris-acetate EDTA (TAE) buffer by a gel electrophoresis system (Shokr et al., 2023). Electrophoresis was performed at a constant 80V for 30min. The amplified product was visualized as a single compact band of expected size under UV light and documented by the Samsung Note4 smartphone.

DNA sequencing for the amplified genes

The amplified PCR products were sent to Solgent Co Ltd (South Korea) for gel purification and sequencing. The amplified PCR products amplified by the universal primer were sequenced using forward and reverse primers. Sequencing was performed using BigDye® Terminator v3.1 Cycle Sequencing Kits and model ABI 3730XL DNA Analyzer (Applied Biosystems, Foster City, CA, USA).

Sequence alignment and phylogenetic analysis

The resulting sequences were quality trimmed and assembled in Geneious software; consequently, the trimmed sequences were identified by search in the basic local alignment Nucleotide Blast tool (BLASTn) of the GenBank database (http://www.ncbi.nlm.nih.gov/BLAST). Nucleotide sequences obtained, together with corresponding sequences retrieved from the GenBank, were aligned using MAFFT alignment (Katoh and Standley, 2013). Phylogenetic trees were constructed using MEGA-X software and the Neighbor-joining method (Saitou and Nei, 1987), employing the Tamura-Nei Model (Tamura and Nei, 1993). The trees were assessed using 1000 bootstrap replicates.

Inoculum preparation

Bacterial cultures stocks were streaked on nutrient agar medium and incubated for 24 h at 37 °C, then one colony of each isolate was inoculated in 5ml nutrient broth in sterile test tubes for 16h, at 37 °C in a shaking incubator (Model: PT. DAIHAN LABTECH) with 150 rpm. 1ml was transferred to 10 ml of sterile nutrient broth medium. After incubation at 37 °C until the cultures reached the mid-exponential growth phase:(OD600 = 0.5), the cultures were diluted with
sterile physiological saline solution (0.85%, sodium chloride) until the turbidity of each suspension was adjusted to match 0.5 McFarland standard (OD600 = 0.132), corresponding to a bacterial density of about 10⁵ CFU/ml (Close et al., 2012).

**Essential oil emulsion preparation**

Tween 80 (1% v/v) was used as an emulsifier for preparing emulsions of the essential oils in nutrient broth medium with a concentration of 55 µl/ml. Tween 80 was sterilized using 0.22µm Millipore filter (Kpadonou et al., 2022).

**Antimicrobial activity test**

The antimicrobial activity of the essential oils (EO) derived from the selected plants was assessed using the agar well diffusion technique. In this method, 0.2 ml of each bacterial suspension was inoculated onto Mueller-Hinton agar medium (Britania) and poured into Petri dishes. Wells with a diameter of 9 mm were created using a sterile cork borer, and 150 µl of undiluted oil was added to each well. As controls, Penicillin (10 µg/disk), Ampicillin (10 µg/disk), and Amoxicillin (25 µg/disk) were used. The diameter of the inhibition zones was measured after incubation for 24 hours at 37°C (Balouiri et al., 2016). This assay was conducted in triplicate, and the mean value of the results was calculated and reported.

**Minimum inhibitory concentration (MIC)**

The minimum inhibitory concentration (MIC) assay was conducted to determine the lowest concentration of the essential oil required to inhibit the growth of the tested bacteria. Initially, 1 ml emulsion of the essential oil was prepared. Serial dilutions of the emulsion were then performed in nutrient broth (NB) medium using sterile test tubes, resulting in final concentrations ranging from 0.075 to 18.33 µl/ml. To each diluted sample, a bacterial suspension with a final concentration of 10⁷ CFU/ml was added. The samples were incubated at 37°C for 24 hours in a shaking incubator set at 150 rpm (Huang et al., 2021). In this assay, control groups were included. These controls consisted of culture medium (NB) only, culture medium (NB) with the bacterial suspension, and NB containing 1% of tween 80 along with the bacterial suspension. To assess bacterial growth, turbidity was measured compared to the control turbidity using a spectrophotometer (Unico, model 7200, made in the USA) at a wavelength of 600 nm (D’Aquila et al., 2022). The MIC was determined as the lowest concentration of the essential oil at which no visible growth (turbidity) of the tested bacteria was observed (Huang et al., 2021). This assay was performed in triplicate to ensure the reliability of the results.

**The potential antibacterial activity of the extracted essential oils**

This assay determined the investigated essential oil had bactericidal or bacteriostatic properties on the tested bacteria. When the bacteria failed to grow on Nutrient broth (NB) and failed to grow again on Nutrient agar (NA) (NEOGEN Culture Medium), even after removing the effect of the essential oil and permanently killed, the effect is bactericidal, but when the bacteria failed to grow on NB and return to grow on NA after removing the effect of essential oil and temporarily stopped from growing, it means bacteriostatic (Smith et al., 1998). This test depended on the previous assay MIC. From the tubes where the bacteria failed to grow and were inhibited at MIC concentration, 50µl was removed, cultured on NA, and incubated at 37°C for 24h for the tested bacteria.

**Statistical analysis**

Results are reported as the mean of three replicates ± standard error (SE). Data were subjected to one-way ANOVA using SPSS (ver. 26). Mean separation was performed using Duncan’s multiple ranges at P<0.05 (Kleinbaum et al., 2013).

**RESULTS**

**Identification of bacterial isolates**

The bacterial isolates were identified using morphological and biochemical characteristics, as shown in Tables (1) which resulted in the identification of two *Bacillus* spp (B1, S1), *Staphylococcus coagulase* negative spp (L1), *Serratia* spp (S3), and *micrococcus* spp (L3).

**Molecular identification of the bacterial isolates using 16S rRNA gene analysis**

Molecular identification revealed that the bacterial isolates are *Bacillus cereus*, *Staphylococcus saprophyticus*, *Lysinibacillus fusiformis*, *Kocuria rhizophila*, and *Serratia liquefaciens* for the bacterial isolates (B1, L1, S1, L3, and S3, respectively) with similarity of 100% then recorded with accession numbers listed in Table (2). Phylogenetic relationships of the isolated strains are shown in Figures (1), (2), and (3). The neighbor-joining method was employed using the maximum composite likelihood bootstrapping with 1000 replicates.

**The water content of the selected plant species**

The water content, moisture content, and succulent values of the three selected medicinal plants are presented in Table (3). *Cupressus macrocarpa* displayed the highest water content, with a moisture content of 54.62%. Conversely, *Eucalyptus citriodora* exhibited the lowest water content, with a moisture content of 38.94%. The succulent values ranged from 1.36 to 2.20 across the selected plant species.

**GC/MS analysis of the extracted essential oils**

Gas chromatography-mass spectrometry (GC/MS) analysis of the medicinal plant tested was employed to identify the constituents of the extracted essential oils. Gas chromatography-mass spectrometry (GC/MS) analysis was performed to identify the constituents of the extracted essential oils from the medicinal plant samples. The results of the GC/MS analysis for each plant are summarized in Tables (4), (5), and (6). Table (4) displays the analysis of *C. macrocarpa* essential oil, revealing the presence of nineteen bioactive compounds, which accounted for 100% of the total oil composition. The major components identified were terpinene-4-ol (32.37%) and citronellol (29.29%), followed by isopulegol (7.06%) and camphor (6.21%) (Fig.4A). However, in the essential oil of *S. terebinthifolius*, a total of twenty compounds were identified.
Table (1): Morphological and biochemical characteristics used for identification of bacterial isolates.

<table>
<thead>
<tr>
<th>Characterization</th>
<th>Tested bacterial isolates</th>
</tr>
</thead>
<tbody>
<tr>
<td></td>
<td>L1</td>
</tr>
<tr>
<td>Macromorphology</td>
<td></td>
</tr>
<tr>
<td>On NA medium</td>
<td></td>
</tr>
<tr>
<td>Shape</td>
<td>Circular</td>
</tr>
<tr>
<td>Color</td>
<td>White</td>
</tr>
<tr>
<td>Opaque</td>
<td>Opaque</td>
</tr>
<tr>
<td>Margins</td>
<td>Entire</td>
</tr>
<tr>
<td>Elevation</td>
<td>convex</td>
</tr>
<tr>
<td>Micromorphology</td>
<td></td>
</tr>
<tr>
<td>Shape</td>
<td>Cocci</td>
</tr>
<tr>
<td>Aggregation</td>
<td>Cluster</td>
</tr>
<tr>
<td>Ability to Gram stain</td>
<td>+</td>
</tr>
<tr>
<td>Spore formation</td>
<td>-</td>
</tr>
<tr>
<td>Pigment Production</td>
<td>-</td>
</tr>
<tr>
<td>Biochemical Test</td>
<td></td>
</tr>
<tr>
<td>Catalase</td>
<td>+</td>
</tr>
<tr>
<td>Coagulase</td>
<td>-</td>
</tr>
<tr>
<td>Voges Proskauer</td>
<td>-</td>
</tr>
<tr>
<td>Indole production</td>
<td>-</td>
</tr>
<tr>
<td>Oxidase test</td>
<td>ND</td>
</tr>
<tr>
<td>Starch hydrolysis</td>
<td>-</td>
</tr>
<tr>
<td>H2S production</td>
<td>-</td>
</tr>
<tr>
<td>Urea hydrolysis</td>
<td>+</td>
</tr>
<tr>
<td>Citrate utilization</td>
<td>-</td>
</tr>
<tr>
<td>Esculin hydrolysis</td>
<td>ND</td>
</tr>
<tr>
<td>Sugar Fermentation</td>
<td></td>
</tr>
<tr>
<td>Mannitol</td>
<td>+</td>
</tr>
<tr>
<td>Lactose</td>
<td>+</td>
</tr>
<tr>
<td>Ability to grow on 5% NaCl</td>
<td>+</td>
</tr>
</tbody>
</table>

+, positive reaction result; -, negative reaction result; ND, not detected.

Table (2): Molecular Identification of foodborne bacterial isolates, recovered from different food sources, using 16S rRNA sequencing analysis.

<table>
<thead>
<tr>
<th>Isolate code</th>
<th>Source of isolation</th>
<th>Sequence length (bp)</th>
<th>BLASTn result</th>
<th>Final identification</th>
<th>Accession number</th>
</tr>
</thead>
<tbody>
<tr>
<td>B1</td>
<td>Beef burger</td>
<td>797</td>
<td><em>Bacillus cereus</em></td>
<td>100 Bacillus cereus DU-BST</td>
<td>OQ380712</td>
</tr>
<tr>
<td>L1</td>
<td>Beef luncheon</td>
<td>1380</td>
<td><em>Staphylococcus saprophyticus</em></td>
<td>100 Staphylococcus saprophyticus DU-LA, Lysinibacillus fusiformis DU-SST</td>
<td>OQ071703</td>
</tr>
<tr>
<td>S1</td>
<td>Sausage</td>
<td>1377</td>
<td><em>Lysinibacillus fusiformis</em></td>
<td>100 Lysinibacillus fusiformis DU-SST</td>
<td>OQ071701</td>
</tr>
<tr>
<td>L3</td>
<td>Beef luncheon</td>
<td>1375</td>
<td><em>Kocuria rhizophila</em></td>
<td>100 Kocuria rhizophila DU-Yell</td>
<td>OQ071700</td>
</tr>
<tr>
<td>S3</td>
<td>Sausage</td>
<td>738</td>
<td><em>Serratia liquefaciens</em></td>
<td>100 Serratia liquefaciens DU-S3</td>
<td>OQ071699</td>
</tr>
</tbody>
</table>
Figure (1): Phylogenetic dendrogram of *B. cereus* (DU-Bst), *Lysinibacillus fusiformis* (DU-SST), and *Staphylococcus saprophyticus* (DU-LA) strains, along with related species, based on 16s rRNA gene sequence alignment. The neighbor-joining was performed using the maximum composite likelihood bootstrapping (1000 replicates).
Figure (2): Phylogenetic dendrogram of *K. rhizophila* (DU-yell) strain along with related species based on 16S rRNA gene sequence alignment. The neighbor-joining was performed using the maximum composite likelihood bootstrapping (1000 replicates).

Figure (3): Phylogenetic dendrogram of *S. liquefaciens* (DU-S3) strain along with related species based on 16S rRNA gene sequence alignment. The neighbor-joining was performed using the maximum composite likelihood bootstrapping (1000 replicates).
Essential Oils against Foodborne Bacteria: Medicinal Plant Potential

representing 100% of the total oil (Fig. 4B).

The main component was α-phellandrene, representing 44.35% of the composition, followed by o-cymene (10.42%) and limonene (6.44%) (Table 5). On the other hand, Eucalyptus citriodora essential oil contained twenty six bioactive compounds, representing 100% of the total oil composition (Fig. 4C). The major components in Eucalyptus citriodora oil were identified as Sabinene (24.24%), α-phellandrene (13.5%), α-eudesmol (7.03%), Spathulenol (6.41%), and o-Cymene (6.09%) (Table 6).

Antibacterial activity of the essential oils

As presented in Figure (5), the essential oil of C. macrocarpa inhibited B. cereus, showing the highest inhibition zone of 55.7±1.2 mm, and exhibited resistance to the tested antibiotics compared to other tested bacteria. However, S. saprophyticus showed inhibition zones of 22.7±1.2 and 31.97±0.55 mm L. fusiformis, 17.5±0.74, and 17.3±0.88 mm for K. rhizophila, and S. liquefaciens, respectively. S. terebinthifolius showed a higher effect against S. saprophyticus with a 24.97±0.84 mm inhibition zone, followed by K. rhizophila and L. fusiformis with inhibition zones of 23.4±1.32 and 23.4±0.17 mm, respectively.

B. cereus and S. liquefaciens showed less sensitivity for S. terebinthifolius as it affected B. cereus with an inhibition zone of 13.8±0.6 and 13.3±0.33 mm for S. liquefaciens. For E. citriodora EO, B. cereus showed the highest inhibition zone of 32.5±0.9 mm,

Table (3): Water content in the selected medicinal plants

<table>
<thead>
<tr>
<th>Medicinal Plant species tested</th>
<th>Measured parameters</th>
<th>Water content (g g⁻¹ DM)</th>
<th>Moisture %</th>
<th>Succulent value</th>
</tr>
</thead>
<tbody>
<tr>
<td>C. macrocarpa</td>
<td></td>
<td>8.09</td>
<td>54.62</td>
<td>2.20</td>
</tr>
<tr>
<td>S. terebinthifolius</td>
<td></td>
<td>6.62</td>
<td>44.90</td>
<td>1.82</td>
</tr>
<tr>
<td>E. citriodora</td>
<td></td>
<td>5.03</td>
<td>38.49</td>
<td>1.63</td>
</tr>
</tbody>
</table>


<table>
<thead>
<tr>
<th>Peak</th>
<th>Compound name</th>
<th>Chemical Classification</th>
<th>Retention time (RT, min)</th>
<th>Molecular Formula</th>
<th>Molecular weight (M.wt)</th>
<th>Area %</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>α-thujene</td>
<td>Monoterpene</td>
<td>8.025</td>
<td>C₁₀H₁₆</td>
<td>136</td>
<td>0.17</td>
</tr>
<tr>
<td>2</td>
<td>sabine</td>
<td>Monoterpene</td>
<td>9.233</td>
<td>C₁₀H₁₆</td>
<td>136</td>
<td>4.31</td>
</tr>
<tr>
<td>3</td>
<td>(-)-β-pinene</td>
<td>Monoterpene</td>
<td>9.776</td>
<td>C₁₀H₁₆</td>
<td>136</td>
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</tr>
<tr>
<td>4</td>
<td>α-terpine</td>
<td>Monoterpene</td>
<td>10.486</td>
<td>C₁₀H₁₆</td>
<td>136</td>
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</tr>
<tr>
<td>5</td>
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<td>Monoterpene</td>
<td>10.577</td>
<td>C₁₀H₁₄</td>
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</tr>
<tr>
<td>6</td>
<td>limonene</td>
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<td>C₁₀H₁₆</td>
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<tr>
<td>7</td>
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<tr>
<td>8</td>
<td>cis-sabinene hydrate</td>
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<td>11.951</td>
<td>C₁₀H₁₇O</td>
<td>154</td>
<td>0.88</td>
</tr>
<tr>
<td>9</td>
<td>α-terpinolone</td>
<td>Monoterpene</td>
<td>12.557</td>
<td>C₁₀H₁₆</td>
<td>136</td>
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<td>10</td>
<td>trans sabine hydrate</td>
<td>Oxygenated monoterpene</td>
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<td>196</td>
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<tr>
<td>12</td>
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<td>13.845</td>
<td>C₁₀H₁₉O</td>
<td>152</td>
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<td>Oxygenated monoterpene</td>
<td>13.976</td>
<td>C₁₀H₁₉O</td>
<td>154</td>
<td>1.13</td>
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<td>Oxygenated monoterpene</td>
<td>15.052</td>
<td>C₁₀H₁₉O</td>
<td>154</td>
<td>32.37</td>
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<tr>
<td>17</td>
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<td>154</td>
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<td>Oxygenated monoterpene</td>
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<td>C₁₀H₂₀O</td>
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80
Minimum inhibitory concentration (MIC)

The MIC for the studied bacterial strains, when treated with *C. macrocarpa* EO, was 0.17 mg/ml for both *S. saprophyticus* and *L. fusiformis*, 0.06 mg/ml for *B. cereus*, 1.5 mg/ml for both *K. rhizophila* and *S. liquefaciens*. The MIC of *S. terebinthifolius* to *B. cereus* and *K. rhizophila* was 2.0 mg/ml, whereas the MICs of *S. saprophyticus* and *L. fusiformis* were 0.68 mg/ml. All investigated bacterial strains showed a similar MIC value for *E. citriodora*, 0.2 mg/ml. The MIC of *S. liquefaciens* to SCHEO was 2.0 and 1.77 mg/ml for both *K. rhizophila* and *S. terebinthifolius*, respectively. The MIC of *B. cereus* was 2.0 mg/ml, whereas the MICs of *S. saprophyticus* and *L. fusiformis* were 0.06 mg/ml. For *B. cereus*, the effect was observed on the tested bacterial strains at the higher concentration of tween 80 (1% v/v).

The potential antibacterial activity of the essential oils

*C. macrocarpa* EO showed a bactericidal effect against *S. saprophyticus* and *S. liquefaciens* as they were completely killed after treatment and failed to grow again. While, for the tested *B. cereus*, the effect was observed on the tested bacterial strains at the higher concentration of tween 80 (1% v/v).

Table (5): Phytochemical composition of essential oils extracted from *Schinus terebinthifolius* using Gas Chromatography/Mass Spectrometry (GC/MS) Analysis.

<table>
<thead>
<tr>
<th>Peak No.</th>
<th>Compound name</th>
<th>Chemical Classification</th>
<th>Retention time (RT, min)</th>
<th>Molecular Formula</th>
<th>Molecular weight (Mwt)</th>
<th>Area %</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>α-pinene</td>
<td>Monoterpene</td>
<td>8.203</td>
<td>C\textsubscript{10}H\textsubscript{16}</td>
<td>136</td>
<td>4.70</td>
</tr>
<tr>
<td>2</td>
<td>(-)-β-pinene</td>
<td>Monoterpene</td>
<td>9.776</td>
<td>C\textsubscript{10}H\textsubscript{16}</td>
<td>136</td>
<td>1.28</td>
</tr>
<tr>
<td>3</td>
<td>α-phellandrene</td>
<td>Monoterpene</td>
<td>10.125</td>
<td>C\textsubscript{10}H\textsubscript{16}</td>
<td>136</td>
<td>44.35</td>
</tr>
<tr>
<td>4</td>
<td>o-cymene</td>
<td>Monoterpene</td>
<td>10.577</td>
<td>C\textsubscript{10}H\textsubscript{14}</td>
<td>134</td>
<td>10.42</td>
</tr>
<tr>
<td>5</td>
<td>sabinene</td>
<td>Monoterpene</td>
<td>10.789</td>
<td>C\textsubscript{10}H\textsubscript{16}</td>
<td>136</td>
<td>3.10</td>
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<tr>
<td>6</td>
<td>limonene</td>
<td>Monoterpene</td>
<td>10.846</td>
<td>C\textsubscript{10}H\textsubscript{16}</td>
<td>136</td>
<td>6.44</td>
</tr>
<tr>
<td>7</td>
<td>α-terpinolene</td>
<td>Monoterpene</td>
<td>12.563</td>
<td>C\textsubscript{10}H\textsubscript{16}</td>
<td>136</td>
<td>1.87</td>
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<tr>
<td>8</td>
<td>terpinen-4-ol</td>
<td>Oxygenated monoterpen</td>
<td>15.035</td>
<td>C\textsubscript{10}H\textsubscript{16}O</td>
<td>154</td>
<td>1.45</td>
</tr>
<tr>
<td>9</td>
<td>α-terpinol</td>
<td>Oxygenated monoterpen</td>
<td>15.372</td>
<td>C\textsubscript{10}H\textsubscript{16}O</td>
<td>154</td>
<td>0.56</td>
</tr>
<tr>
<td>10</td>
<td>citronellol</td>
<td>Oxygenated monoterpen</td>
<td>16.534</td>
<td>C\textsubscript{10}H\textsubscript{20}O</td>
<td>156</td>
<td>4.61</td>
</tr>
<tr>
<td>11</td>
<td>5-isopropenyl-2-methyl-7-oxabicyclo[4.1.0]heptan-2-ol</td>
<td>Oxygenated monoterpen</td>
<td>16.643</td>
<td>C\textsubscript{10}H\textsubscript{16}O\textsubscript{2}</td>
<td>168</td>
<td>4.78</td>
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<tr>
<td>12</td>
<td>cis-p-mentha-1(7),8-dien-2-ol bicyclo[3.1.0]hexan-3,6-diol</td>
<td>Oxygenated monoterpen</td>
<td>18.662</td>
<td>C\textsubscript{10}H\textsubscript{16}O</td>
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<td>2.38</td>
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<tr>
<td>13</td>
<td>1-(1-methylthyl)-1,3-dimethylen-1-(1-methylthyl)-1,3-dimethyl-1,3,6,8-dien-2-ol</td>
<td>Oxygenated monoterpen</td>
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<td>Sesquiterpen</td>
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<td>204</td>
<td>1.52</td>
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<td>15</td>
<td>caryophyllene</td>
<td>Sesquiterpen</td>
<td>21.77</td>
<td>C\textsubscript{15}H\textsubscript{24}</td>
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<td>3.05</td>
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<tr>
<td>16</td>
<td>β-cubebene</td>
<td>Sesquiterpen</td>
<td>23.229</td>
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<tr>
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<td>Sesquiterpen</td>
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<tr>
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<td>Oxygenated Sesquiterpen</td>
<td>25.357</td>
<td>C\textsubscript{15}H\textsubscript{24}O</td>
<td>220</td>
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</table>

Total identified: 100.00
Table (6): Phytochemical composition of essential oils extracted from *Eucalyptus citriodora* using Gas Chromatography/Mass Spectrometry (GC/MS) Analysis.

<table>
<thead>
<tr>
<th>Peak No.</th>
<th>Compound name</th>
<th>Chemical classification</th>
<th>Retention time (RT, min)</th>
<th>Molecular Formula</th>
<th>Molecular weight</th>
<th>Area %</th>
</tr>
</thead>
<tbody>
<tr>
<td>1</td>
<td>α-thujene</td>
<td>Monoterpene</td>
<td>8.031</td>
<td>C_{10}H_{16}</td>
<td>136</td>
<td>0.88</td>
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<tr>
<td>3</td>
<td>β-pinene</td>
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<td>C_{10}H_{16}</td>
<td>136</td>
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</tr>
<tr>
<td>4</td>
<td>α-phellandrene</td>
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<td>154</td>
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<td>C_{3}H_{9}O</td>
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<tr>
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<td>C_{3}H_{9}O</td>
<td>154</td>
<td>2.56</td>
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<td>C_{13}H_{24}</td>
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</tr>
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<tr>
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<td>β-eudesmol</td>
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<td>C_{13}H_{24}</td>
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<tr>
<td>26</td>
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</tbody>
</table>

Table (7): Antibacterial activity of the extracted essential oils verses three standard traditional antibiotics (Penicillin, Amoxicillin, and Ampicillin).

<table>
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<tr>
<th>Tested materials</th>
<th>Tested Bacteria</th>
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<tr>
<td></td>
<td><em>B. cereus</em></td>
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<tr>
<td><strong>Essential Oil</strong></td>
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</tr>
<tr>
<td><em>C. macrocarpa</em></td>
<td></td>
</tr>
<tr>
<td></td>
<td>55.70 ± 1.2</td>
</tr>
<tr>
<td><em>S. terebinthinifolius</em></td>
<td></td>
</tr>
<tr>
<td></td>
<td>13.77 ± 0.6</td>
</tr>
<tr>
<td><strong>E. citriodora</strong></td>
<td></td>
</tr>
<tr>
<td></td>
<td>32.50 ± 0.9</td>
</tr>
<tr>
<td><strong>Standard Antibiotic</strong></td>
<td></td>
</tr>
<tr>
<td>Penicillin</td>
<td>0.00 ± 0</td>
</tr>
<tr>
<td>Amoxicillin</td>
<td>0.00 ± 0</td>
</tr>
<tr>
<td>Ampicillin</td>
<td>0.00 ± 0</td>
</tr>
</tbody>
</table>

1 Essentials oil extracted from selected three medicinal plants; ††, Standard traditional antibiotic used as a reference.
Figure (4): GC/MS Chromatogram of the essential oils extracted from different medicinal plants. A, *C. Macrocarpa*; B, *S. terebinthifolius*, and C, *E. citriodora*. 
of C. macrocarpa oil was bacteriostatic as it lost 95.9% from its normal growth. Treatment of K. rhizophila and L. fusiformis showed that K. rhizophila lost 98.58% of its normal growth, and L. fusiformis lost 97.85% of its normal growth, meaning the effect was static against them.

The activity of S. terebinthifolius was bacteriostatic on the tested Gram-positive bacteria. K. rhizophila lost 98.7% of its normal growth, B. cereus lost 94% of its normal growth, and L. fusiformis lost 99% of its normal growth. S. saprophyticus lost 98.57% of its normal growth. The bacteriostatic activity was observed on the tested Gram-positive bacteria when treated with E. citriodora and allowed to grow again. K. rhizophila lost 91.6% of its normal growth, B. cereus lost 94.5% of its normal growth, L. fusiformis lost 97.20% of its normal growth, L. fusiformis lost 97.20% of its normal growth, and S. saprophyticus lost 85.20% of its normal growth. S. terebinthifolius and E. citriodora showed a bacteriostatic effect on S. liquefaciens as it lost 96 and 98.3%, respectively, from its normal growth after being treated compared with its normal growth without treatment by oils.

**DISCUSSION**

This study studied the antibacterial activity of essential oils from some medicinal plants against some foodborne bacteria to be used as alternative natural food preservatives. Using GC/MS analysis, the essential oil of C. macrocarpa contained nineteen bioactive compounds. Terpinen-4-ol (32.37%), citronellol (29.29%), isopulegol (7.06%), camphor (6.21%), and sabine (4.31%) constitute the majority of the oil. Badawy and Abdelgaleil, (2014) reported that the major component in the essential oil of Egyptian C. macrocarpa leaves was terpinen-4-ol and represented by 20.29%. Also, it was reported that α-terpineol (19.01%) was the major constituent in C. macrocarpa (Saad et al., 2017), while Salem et al. (2018) reported that the main constituents of the EO obtained from Egyptian Cupressus macrocarpa were terpinen-4-ol (23.7%), α-phellandrene (19.2%), and citronellol (17.3%). The major compounds identified in S. terebinthifolius were α-phellandrene (44.35%), α-cymene (10.42%), and limonene (6.44%). The chemical composition of S. terebinthifolius was studied in the literature by Santana et al., (2012), who reported that β-longipinene (8.1%), germacrene D (23.8%), bicyclo germacrene (15.0%) and the monoterpenes α-pinene (5.7%) and β-pinene (9.1%) were the main components of the essential oil extracted from leaves of S. terebinthifolius. Belhoussaine et al., (2022) reported that the majority of the compounds present in the essential oil of S. terebinthifolius leaves were limonene 23.22%, spathulenol 14.34%, γ-terpinene 9.45%, and β-ocimene 13.32%. For Eucalyptus citriodora EO, Sabine (24.24%), α-phellandrene (13.5%), α-eudesmol (7.03%), (-)-Spathulenol (6.41%), and α-Cymene (6.09%) were found to be the major constituents in E. citriodora. Salem et al., (2018) reported that α-citronellal (56%) was the primary constituent of E. citriodora EO. The variation in chemical composition between the same plant species is attributed to different factors. These factors were cultivation type, vegetative stage, plant season (Ghasemzadeh et al., 2016), extraction method, harvesting time, plant part (Mesomo et al., 2013) and age of plant has great effect on its essential oil biological activity and chemical composition (Farias et al., 2023).

C. macrocarpa EO exhibited antibacterial activity against all the tested bacterial strains, with MIC values ranging from 0.06 to 1.5 mg/ml. Bacillus cereus showed the highest inhibition zone of 55.7 mm, with MIC of 0.06 mg/ml. According to Salem et al., (2018), Staphylococcus aureus and Bacillus cereus were both inhibited by the EO from C. macrocarpa with MICs of 0.31 and 0.12 mg/ml, respectively, and the bactericidal effect was at 0.2 and 0.41 mg/ml respectively. Due to variations in the essential oil's chemical composition and its bioactive components, as mentioned before, the results varied.

The component of C. macrocarpa essential oil showed an antibacterial effect, e.g., α-phellandrene (Zhang et al., 2017), camphor and limonene (Han et al., 2019), γ-terpinene (Giweli et al., 2012). Terpenoids are biologically active compounds with

### Table (8): Minimal inhibition concentration (mg ml⁻¹) assed for essential oil extracted from different selected medicinal plants against foodborne bacteria isolated from spoiled food.

<table>
<thead>
<tr>
<th>Plant essential oil extracted</th>
<th>B. cereus</th>
<th>S. saprophyticus</th>
<th>L. fusiformis</th>
<th>K. rhizophila</th>
<th>S. liquefaciens</th>
</tr>
</thead>
<tbody>
<tr>
<td>C. macrocarpa</td>
<td>0.06</td>
<td>0.17</td>
<td>0.17</td>
<td>1.50</td>
<td>1.50</td>
</tr>
<tr>
<td>S. terebinthifolius</td>
<td>2.00</td>
<td>0.68</td>
<td>0.68</td>
<td>2.00</td>
<td>2.00</td>
</tr>
<tr>
<td>E. citriodora</td>
<td>0.20</td>
<td>0.20</td>
<td>0.20</td>
<td>0.20</td>
<td>1.77</td>
</tr>
</tbody>
</table>
higher effects on the resistance to pathogens (Sharma et al., 2019). Zhang et al., (2018) reported that terpinen-4-ol showed antibacterial activity against S. aureus and Streptococcus agalactiae. Huang et al., (2021) reported that terpinen-4-ol, α-terpineol, and δ-terpineol the three isomeric terpineols isolated from Cinnamomum longepaniculatum leaf oil by rupturing the cell wall and membrane of bacteria and resulting in cell death, have the potential to be used as antibacterial compounds. Monoterpenoids interfere with microorganisms’ physiological and biochemical processes to develop and multiply (Pandey et al., 2016).

The essential oil from S. terebinthifolius (SCHEO) showed antibacterial activity. S. saprophyticus showed the highest inhibition zone diameter of 24.97 mm, but low activity was observed against the Gram-negative isolate, S. liquefaciens showed an inhibition zone diameter of 13.3 mm, and this agrees with that reported by El-Massry et al. (2009) as the Egyptian S. terebinthifolius showed inhibitory activity against the tested, bacteria like Pseudomonas aeruginosa, Staphylococcus aureus, and Escherichia coli. Low activity was observed against E. coli, and this may be due to lipopolysaccharides in the outer membrane of the Gram-negative bacteria, which make them resistant to external agents such as antibiotics, detergents, and hydrophilic dyes (Negi and Jayaprakasha, 2003). Gonzalez et al. (2004) reported that α-phellandrene and limonene were the main components of Schinus molle EO, which showed high activity against Bacillus cereus. Several EOs contain the very common cyclic monoterpene α-phellandrene, which exhibits a wide range of biological activity (Radice et al., 2022). The effectiveness and reduction of any pathogenic microorganism’s resistance evolution are due to the synergism between the aromatic plant components (Elshafie et al., 2022). From our results, the investigated Gram-positive bacteria were found to be more sensitive to Eucalyptus essential oil than the tested Gram-negative. The essential oil derived from E. citriodora has antimicrobial properties (Salem et al., 2018; Barbosa et al., 2016).

From our findings, SCHEO exhibited antibacterial activity against the tested bacteria with a MIC value of 0.68 mg/ml for L. fusiformis and S. saprophyticus, but it was 2.0 mg/ml for the other tested bacteria. According to El-Massry et al., (2009), SCHEO showed antibacterial activity against Staphylococcus aureus, Pseudomonas aeruginosa, and Escherichia coli with MIC 0.80, 0.80, and 1.10 mg/ml. For Eucalyptus oil, the MIC for Gram-positive bacteria (0.20 mg/ml) was lower than that for Gram-negative bacteria (1.77 mg/ml). Salem et al. (2018) reported that EO from E. citriodora leaves showed antibacterial activity with MIC values ranging from 0.06 to 0.20 mg/ml.

The antibacterial effects of natural agents may be related to some reasons, such as the inhibition of biofilm formation (Latifah-Munirah et al., 2015; Molina Bertrán, 2022), the rupture of the cell wall and the cell membrane (Cui et al., 2018; Wongsawan et al., 2019), the influence on DNA replication (Rajkowska et al., 2017). Essential oils’ structure, functional groups, and composition affect their antibacterial properties (Gunasena et al., 2022). Due to EOs’ high concentration of lipophilic compounds, which allowed them to adhere to bacterial cell wall surfaces and membrane structures and damage their integrity, cell lysis resulted (Brozyna et al., 2021). Because of their low molecular weight and highly lipophilic nature, terpenes and terpenoids can disrupt cell membranes, kill cells, or prevent the germination and sporulation of fungi that cause food spoilage. This property may be the source of an essential oil’s antimicrobial activity. A previous study of in vitro investigations suggests that terpenes
and terpenoids perform inadequately as antimicrobials when used separately from the whole EO (Tian et al., 2011). Compared to Gram-positive bacteria, Gram-negative bacteria are more resistant to hydrophobic antibiotics as Gram-negative bacteria have a surface that acts as a barrier to the entrance of macromolecules and hydrophobic substances into the bacteria cell membrane (Teerarak and Laosinwattana, 2019) as it has hydrophilic lipopolysaccharides (LPS) on their outer membranes, which acting as a barrier to hydrophobic substances, causing them to have a higher tolerance for hydrophobic antimicrobial compounds such those found in essential oils (Nikaido, 2003) and that's agreed with our results.

There have been reports that terpenes and terpenoid compounds have bactericidal properties. Antimicrobial substances must penetrate or rupture the bacterial plasma membrane to kill bacteria. Any tiniest change to the cell membrane's stability and structural integrity could harm the bacteria's regular metabolic processes, which would reduce the viability of the cell (Patra et al., 2015). Terpenes, when interacting with cells, affect several metabolic processes, particularly those associated with energy production and cell membrane transport mechanisms, as reported by Aleksić and Knezevic (2014). According to several studies, most essential oil bioactive components kill bacteria by rupturing their cell walls and membranes (Diao et al., 2014; Xu et al., 2017). The active compounds in essential oils could prevent the synthesis of nucleic acids, proteins, and other macromolecular compounds that would inhibit bacterial growth (Diao et al., 2014). Essential oils can interfere with pH equilibrium, homeostasis, and the integrity of the cell membrane of microorganisms (Lambert et al., 2001). Additionally, they can affect the depolarization of cell membranes (Mecom et al., 2017) and interfere with their permeability (Kumar et al., 2019).

**CONCLUSION**

In conclusion, the extracted essential oils from C. macrocarpa, S. terebinthifolius, and E. citriodora have demonstrated remarkable potential as natural antimicrobial agents. Through rigorous experimentation, these essential oils have shown significant antibacterial properties against S. saprophyticus, B. cereus, L. fusiformis, K. rhizophila, and S. liquefaciens, which are common contaminants and spoilage agents in food products. The emergence of these essential oils as effective bio-preservatives presents a promising avenue for natural alternatives to traditional synthetic food preservatives. By connecting the power of these botanical treasures, it becomes possible to enhance the shelf life of food products while reducing reliance on artificial additives. However, further research is warranted to explore the full potential of these essential oils and their application in the food industry. Factors such as optimal concentrations, formulation techniques, and compatibility with various food matrices should be thoroughly investigated to ensure their safe and effective utilization. This study contributes to our understanding of the antimicrobial properties of essential oils, providing a foundation for a greener and more sustainable approach to food preservation. By utilizing these natural resources, we can strive towards a healthier and safer food industry.

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فاعليّة الزيوت العطرية لثلاثة نباتات طبية كمضادات لبعض البكتيريا المنقولة بواسطة الغذاء

محمود سالم سراج، دعاء عاطف، ريهام أحمد الفيومي
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الخلاصة

تم استخلاص الزيوت العطرية من ثلاث نباتات طبية وهي السرو الليموني، الفلفل البرازيلي، والكافور الليموني باستخدام التقطير المائي وجهاز كليفينجر. وتم تحليل الزيوت باستخدام GC/MS للحصول على مكونات الزيوت. وتم الدراسة على 19 مادة كيماويًا من البكتيريا المستخدمة في التحلي. وتم تحديد القيم النشطة في مكافحتهم من الناحية الجينسي. وتم تقييم الزيوت في مكافحة البكتيريا المختلفة باستخدام الاختبارات المخبرية. وتم اكتشاف مجموعات من الزيوت العطرية النشطة في مكافحة البكتيريا المستخدمة في التحلي. وتم تحديد النتائج المحتملة للتطبيق العملي للزيوت العطرية المستخلصة كمضادات للأمراض المعدية.